Wild Furbearer Management and Conservation in North America

Edited by Tim L. Hiller, Roger D. Applegate, Robert D. Bluett, S. Nicki Frey, Eric M. Gese, and John F. Organ

Chapter 17: Chemical Immobilization of Furbearers

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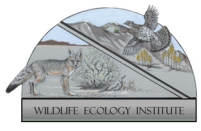

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CHEMICAL IMMOBILIZATION OF FURBEARERS

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Chemical immobilization is the use of approved pharmaceuticals to reduce an animal's movements so that it can be safely handled by humans while reducing the potential for harm to itself. Although many species of furbearers can be physically restrained, this can be very psychologically and physiologically stressful to the animal as well as making tasks such as collecting invasive samples (e.g., blood), attaching radio-transmitters, and measuring for morphometrics more difficult than needed. Most modern immobilizing drugs for furbearers have been in use for decades and have been proven to be extremely safe and reliable. Immobilizing drugs can be used for safe removal of animals from traps; handling, and sampling; anesthetizing furbearers for painful procedures such as tooth extraction, transmitter implants, and wound management; transportation during translocation efforts; and removal of furbearers in problematic areas.

This chapter includes legalities of drug use, drug pharmacology, equipment, and animal medical considerations, and may be considered an update of information provided by Seal and Kreeger (1987). It is intended to give readers an overview of this field without in-depth technical discussions. A more technical and expansive coverage of chemical immobilization can be found in Kreeger et al. (2023). The most substantial changes since Seal and Kreeger (1987) concern the development of potent and antagonizable sedatives that reduce anesthetic doses and combinations of multiple drugs that provide efficacy and antagonist capabilities while increasing safety for both animals and humans.

DRUG POSSESSION AND USE

Acquisition and Possession of Drugs

Canada

Veterinarians must practice according to enforceable rules of conduct or bylaws and codes of ethics, which are developed by each provincial veterinary medical licensing body. Depending on provincial regulations, a veterinarian may be able to prescribe non-controlled drugs for use by a non-veterinarian as long as a valid veterinarian-client-patient relationship has been established. Unfortunately, this does not apply to controlled substances.

Virtually all drugs used for wildlife immobilization in Canada are controlled substances under the 1996 Controlled Drugs and Substances Act (Government of Canada 2020). There are 9 schedules of controlled substances under this Act. Examples of schedules for immobilization drugs commonly used on wildlife species include:

Schedule I: carfentanil, etorphine, fentanil, ketamine, sufentanil, and thiafentanil.

Schedule IV: butorphanol, diazepam, midazolam, and nalbuphine.

Drugs such as azaperone, medetomidine, and xylazine are not covered under this Act, but are still prescription drugs and must be used on or by the order of a licensed veterinarian.

An Experimental Studies Certificate (ESC) issued by the Veterinary Drugs Directorate (VDD) permits a manufacturer to sell drugs to qualified individuals, and controlled drugs to persons holding a Section 56 exemption (see below). A qualified non-veterinarian could be a government employee or legitimate research scientist who has passed a recognized chemical immobilization course and is familiar with the drugs being used. Normally, an ESC is required for every project involving chemical immobilization of wildlife, with projects usually considered ≤6 months in duration. Additionally, criteria have been established by the Veterinary Drugs Directorate and the Drug Strategy and Controlled Substances Program (DSCSP) for authorization to acquire and possess controlled immobilizing agents for wildlife research through an exemption as described in Section 56 of the Act.

Applicants must make a written request to the minister for every project. The DSCSP evaluates these requests and issues authorizations on behalf of the minister. The application must include a detailed protocol or outline of the project. Requirements include that the applicant must: 1) have at least an undergraduate degree in biology, 2) be employed as a wildlife biologist, 3) have a current certificate in CPR, and 4) have successfully completed and passed a course in chemical immobilization. Anyone who purchases, possesses, or uses ketamine (or other controlled substance) must therefore have: 1) an approved ESC or be

authorized under an ESC, and 2) a letter of exemption to use a controlled substance for scientific purposes under Section 56 of the Controlled Drugs and Substances Act.

United States

Conditions for the use of drugs (pharmaceuticals) to sedate or immobilize animals are established by the U.S. Food and Drug Administration (U.S. FDA). U.S. FDA verifies the safety and efficacy of drugs as well as ensures manufacturing quality control. Drug manufacturers must undergo a lengthy and expensive process of drug testing to receive approval from U.S. FDA. This approval includes limits (e.g.., intended species, dose, conditions of use, withdrawal times) for the use of individual drugs to conditions specified on the label. Few drugs have been specifically approved by U.S. FDA for use on wildlife, none of which are furbearers. Any use of these or other drugs on any species not identified on the label is termed extra-label or off-label use.

In addition to being prescription drugs, some of the drugs used for immobilization of wildlife are termed controlled substances. A controlled substance is a drug that is identified in 1 of 5 schedules by the U.S. Drug Enforcement Administration (U.S. DEA). Federal legislation governing the possession of controlled substances is contained in the Controlled Substances Act of 1970. Special regulations govern the recording of use and storage of these drugs (U.S. DEA 2019). The Controlled Substances Act requires an individual to have a U.S. DEA registration number (U.S. DEA Form 224/225) in order to possess controlled substances. Controlled drugs fall into 1 of 5 schedules:

Schedule I: reserved for experimental and abused drugs, such as heroin, marijuana, and lysergic acid diethylamide (LSD); there are no Schedule I drugs used on wildlife.

Schedule II (IIN): includes most of the opioids used for animal immobilization, such as etorphine, fentanyl, sufentanil, and carfentanil, and the opioid antagonist, diprenorphine.

Schedule III (IIIN): includes ketamine and tiletamine-zolazepam, which are used extensively on wildlife.

Schedule IV: includes diazepam, midazolam, alfaxalone, and butorphanol.

Schedule V: includes small, limited quantities of narcotic drugs included in preparations with non-narcotic active medicinal ingredients; there are no Schedule V drugs used on wildlife.

Other drugs used for chemical immobilization of furbearers and that are not listed as controlled substances by the U.S. DEA include most sedatives and tranquilizers (e.g., dexmedetomidine, xylazine), inhalant (gas) anesthetics (e.g., isoflurane, sevoflurane), and adjuvants (e.g., epinephrine, neostigmine).

The Animal Medicinal Drug Use Clarification Act of 1994 (AMDUCA; passed into law in 1996; U.S. FDA 2018) essentially allowed veterinarians to use approved animal and human drugs on wildlife under certain conditions. The AMDUCA makes a specific distinction between food and non-food animals, but for the purposes of this chapter, it is assumed that furbearers are generally not consumed by humans. Extra-label use of animal or human drugs is allowed in non-food animals if the drug is: 1) approved by the U.S. FDA, 2) used by or on the lawful written or oral order of a licensed veterinarian, and 3) used within the context of a valid veterinarian-client-patient relationship.

Biologists may apply for a U.S. DEA registration number, and if approved, may then procure drugs through veterinary product distributors. Technically, however, even though they are in possession of these drugs, they cannot legally use these drugs on animals without veterinary supervision (Clapham et al. 2019). If a biologist uses veterinary prescription drugs without the involvement of a licensed veterinarian, they are in violation of federal regulations. All drugs currently used for the chemical immobilization of wildlife are prescription drugs and must be used by or on the order of a licensed veterinarian. Non-veterinarians can legally use drugs if a valid veterinarian-client-patient relationship is established. That is, the biologist serves as the client and the wild animal serves as the patient. The biologist consults with the veterinarian on the use of the drug to determine the appropriate dose and application. In the U.S., the veterinarian does not have to be on site during the immobilization process, but the veterinarian should be involved in the planning process.

Records on Inventory and Use

Records for individual animal captures are valuable for reviewing efficacy of the drugs and doses, keeping track of samples, determining reasons for adverse reactions, and to conform to federal regulations governing controlled substances. Formats of records are as diverse as the individuals designing them. An example of an individual animal record can be found in Kreeger et al. (2023). An inventory record must also be maintained for all controlled drugs and is subject to audit by the U.S. DEA. Such records should contain the following information, at a minimum, for purchase inventory: 1) type of drug received (e.g., etorphine, ketamine), 2) amount received, 3) date received, and 4) source of drug received (i.e., manufacturer or distributor); and for use inventory: 1) amount used, 2) date used, 3) species on which used, and 4) reason for use.

Amounts of drug used must be reconciled with amounts of drug received. For example, if 50 ml of ketamine was received, then 50 ml of ketamine must be recorded as used or still in inventory. Reasons for use can include not only administration to animals, but also lost darts during capture efforts, accidental spillage, and intentional disposal of unused drug. Records in the U.S. must be maintained for 2 years and inventories should be conducted biannually.

Ordering and Storing Drugs

In the U.S., all Schedule II controlled substances must be ordered using U.S. DEA Form 222 (or its electronic equivalent), which is issued to the holder of the U.S. DEA registration number. This form must be sent to the drug manufacturer or distributor. However, before any drug is shipped, the holder must have approved storage facilities for these drugs. Schedule II controlled

substances must be stored in a safe or steel cabinet equivalent to a U.S. Government Class 5 security container. This usually means a safe weighing >340 kg (750 lb) or a safe that is bolted to the floor with the bolts brazed in such a manner as to prevent tampering. The local U.S. DEA office must then physically inspect the storage container and send their recommendations for approval to the U.S. DEA in Washington, D.C. The U.S. DEA will then notify the manufacturer or distributor that the individual is approved. All other controlled substances must be stored in a secure place with limited access. Regulations regarding drug storage are contained in 21 CFR 1301.75d. For more information regarding drug storage, contact the local U.S. DEA office. Wildlife professionals often transport drugs to field settings within their state, and perhaps across states, but should seek recommendations from their local U.S. DEA (and the appropriate state regulatory) office to ensure compliance.

Expiration Dates

It has long been presumed that an expiration date of a drug (printed on the drug label) was the date determined by a drug manufacturer at which the drug retained ≥90% of its efficacy. This may not be correct. Although it cannot be recommended to use drugs after the expiration date, there is scientific evidence that many drugs remain 100% effective for many years, even decades, after the expiration date (Cantrell et al. 2012, Diven et al. 2015).

Controlled and field trials have been conducted to test the hypothesis that immobilizing and antagonist drugs remain viable after many years (Kreeger et al. 1990*a*; J. Arnemo, Inland Norway University of Applied Sciences, personal communication). Drugs such as etorphine, medetomidine, naloxone, and naltrexone remain effective for 5–10 years past their expiration dates. In support of such findings, Diven et al. (2015) suggested that expiration dates for ketamine be extended by 64 months and naloxone by 77 months. However, care should be taken with bottle condition and number of seal punctures to minimize contamination.

It may be difficult to discard a vial of drugs, which may have been very costly, just because it has reached its expiration date. However, using drugs past their expiration date assumes some risk of liability. Chemical immobilization of animals is unpredictable and relatively uncontrolled. If an expired drug was used to immobilize an animal and a person was injured or property was damaged during the immobilization process, the person that administered the drug and their employer may be held responsible. The use of any expired drugs might best be reserved for animals under controlled settings, such as captivity.

Calculating Drug Doses

Accurate calculation of drug doses is critical to reduce the problems associated with underdosing or overdosing animals. Information required prior to calculating a dose includes: 1) weight of individual animal, 2) drug concentration, and 3) dose (mg of drug/kg of body weight). For those lacking experience with the average weights by sex-age class (male, female; juvenile, adult) of a particular species, either contact someone who has experience or see Chapters 27–66 (Hiller et al. 2024). Most manufacturers provide concentrations (i.e., mg/ml) of their products in mg of drug/ml of solvent. Some drugs are freeze-dried (lyophilized). To prepare a solution of known concentration, calculate backwards from the desired solution to arrive at the volume of solvent to add to the powdered drug. That is, if a drug bottle contains 500 mg of drug and a 100-mg/ml solution is desired, add 5 ml of solvent to the bottle (i.e., 100 mg/ml = 500 mg/5 ml). Ultimately, you want to know what volume of drug to administer to the animal. The formula for this is:

$$\text{Volume of drug administered} = \frac{\text{Body weight} \times \text{Dose}}{\text{Drug concentration}}$$

Consider immobilizing an animal that weighs 10 kg (22 lb) with Drug *X*. The recommended dose of Drug *X* for this animal is 5 mg/kg. Drug *X* is available in a 100-mg/ml solution. First, calculate the total amount (mg) of drug needed to chemically immobilize this animal by multiplying the animal's weight (10 kg) by the recommended dose of drug (5 mg/kg):

$$\text{Drug } X \text{ needed (mg)} = 10 \text{ kg} \times 5 \text{ mg/kg} = 50 \text{ mg}$$

Then calculate the volume of the drug solution to withdraw from the bottle by dividing the dose (i.e., 50 mg) of the drug by the concentration (100 mg/ml) of the drug:

$$\text{Volume (ml) needed} = 50 \text{ mg}/100 \text{ mg/ml} = \text{0.5 ml of Drug solution}$$

Don't trust memory when calculating drug doses unless you consistently use the same drug(s) on the same species. Also, double-check the calculation to determine if the calculation is logical. With user experience, a miscalculated drug volume should trigger a mental alarm.

DRUGS USED FOR CHEMICAL IMMOBILIZATION OF FURBEARERS

The Evolution of Drugs

This chapter is about immobilization, as opposed to tranquilization or anesthesia, of furbearers. Immobilization was a term that initially referred to some of the earliest drugs used to capture animals. These drugs were paralyzing drugs, like succinylcholine. Such drugs, while rendering the animal immobile, did not render the animal unconscious. The next drugs used to capture animals were the barbiturates and the cyclohexanes, both of which were true injectable anesthetics (i.e., induced unconsciousness). In the 1960s, the first of the potent opioids (etorphine) was developed, followed by even more potent opioids (carfentanil, thiafentanil). The opioids, however, seemed to induce a state that was neither paralysis nor anesthesia, but rather induced a state when combined with tranquilizers termed neuroleptanalgesia.

In the past, drugs used for chemical immobilization were considered as primary and secondary immobilants. The primary immobilant (e.g., etorphine, ketamine) was sufficient to induce

immobilization on its own. Secondary immobilants were tranquilizers or sedatives which, when combined with the primary immobilant, resulted in improved immobilization (e.g., use of ketamine-xylazine). By themselves, tranquilizers or sedatives only induced a state of calmness. Such calmness may be profound to the point that the animal may seem to be unconscious and can be safely handled (e.g., administering only medetomidine for foxes [*Vulpes* spp.]). However, if sufficiently stimulated, a tranquilized or sedated animal can arouse and flee, or possibly attack.

Today, there are drug combinations with essentially no primary immobilant. Instead, a mixture of drugs, none of which are individually capable of safely immobilizing a wild animal, are used to induce a state of profound sedation or neuroleptanalgesia. These combinations were developed because of the loss, restrictions, or regulations of several primary immobilants (e.g., carfentanil, thiafentanil). Examples of several recent drug combinations include butorphanol-azaperone-medetomidine (BAM), nalbuphine-azaperone-medetomidine (NAM), and alfaxalone-azaperone-medetomidine (AAM).

Agonistic Drugs

An agonist drug is a chemical that binds to a receptor to produce a biological response. Such drugs include anesthetics, opioids, paralytics, tranquilizers, and other drugs used for the chemical immobilization of wildlife.

Succinylcholine

Succinylcholine is a neuromuscular-blocking drug and was one of the first drugs used for the chemical immobilization of wildlife. Immobilization is characterized by an initial transient rapid firing of the muscles (muscle fasciculations), which is quickly replaced by general paralysis. The order of paralysis is sequential, starting with the jaw, tail, and face, followed by legs and neck, throat, abdomen, intercostal muscles, and diaphragm. Recovery occurs in the reverse order.

Despite its long history of use, succinylcholine is generally inferior to modern drugs. There are two major deficiencies of succinylcholine. One is that it has a very low therapeutic index (an estimation of drug safety), where dose errors of only ±10% can result in either no effect or death. Overdosing results in diaphragmatic paralysis and death by asphyxia. Mortality rates of ≤70% have occurred. The second deficiency is that succinylcholine is virtually devoid of central nervous system (CNS) effects because of its inability to cross the blood-brain barrier. Thus, an animal paralyzed with succinylcholine is conscious, aware of its surroundings, fully sensory, and as such, can feel pain and experience psychogenic stress, yet is physically unable to react. Strictly from the animal's perspective, however, there may be little perceived difference between being chemically paralyzed and physically restrained.

There are some advantages to succinylcholine. It is generally very fast-acting (1–5 min) and the duration of effect is relatively brief (15–30 min). Also, animals that have been administered only succinylcholine, and that have died or been euthanized using physical means (i.e., not other drugs), can be safely consumed by scavengers. Succinylcholine should be used judiciously and only under the most unique circumstances. Nonetheless, succinylcholine is frequently used for immobilization of large animals in U.S. game farms or for euthanasia of furbearers by some wildlife agencies. Succinylcholine is not a controlled substance.

Acepromazine

Acepromazine is a phenothiazine tranquilizer that potentiates analgesic and anesthetic properties of other drugs. It is not a controlled substance, mixes readily with primary immobilants, and is commonly available from most veterinarians. Clinical effects of acepromazine can last 4–8 hours. When combined with ketamine, acepromazine can safely immobilize any furbearing species.

Azaperone

Azaperone is a butyrophenone tranquilizer, which has been reported to counteract opioid respiratory depression in wild animals (Marsboom 1969). It is fast-acting, relatively safe, and not a controlled substance. Its primary use is in BAM or NAM to immobilize some of the larger species of furbearers (e.g., bobcats [*Lynx rufus*], gray wolves [*Canis lupus*]).

Diazepam and Midazolam

Diazepam and midazolam are benzodiazepine tranquilizers used primarily in immobilization of furbearers as anticonvulsant adjuncts to the cyclohexane anesthetics (e.g., ketamine). They are also excellent muscle relaxants, and both drugs are Schedule IV controlled substances. Diazepam is solubilized in 40% propylene glycol, which may produce cardiac arrest if injected too rapidly intravenously (IV). Midazolam is in an aqueous base and does not cause these cardiovascular reactions. Specific antagonists (e.g., flumazenil, sarmazenil) are available that could decrease recovery times. These tranquilizers, combined with ketamine, are extremely safe and effective for the immobilization of smaller species of furbearers. Their relatively low concentrations (5 mg/ml) generally preclude their use in larger species of furbearers, such as gray wolves, because of the high volumes of drug that are necessary.

Xylazine, Detomidine, Medetomidine, and Dexmedetomidine

These potent sedatives are alpha-2 adrenoceptor agonists that can be completely antagonized by use of specific antagonists (chemicals that block or reverse the action of an agonist). They are widely used with primary immobilants (e.g., ketamine, tiletamine) and in multiple-drug combinations. By themselves, they are capable of heavily sedating animals, including furbearers, to the point of relatively safe handling (Baldwin et al. 2008). Sedation of highly excited animals using alpha-2 adrenoceptor agonists alone, however, will be prolonged, if not impossible. If a sedated animal is aroused, eliminating the stimulation will usually result in resedation, recumbency, or both.

When sedating restrained furbearers by using only alpha-2 adrenoceptor agonists, maintain a safe distance from the animal after injection so the animal can calm down. Sedation could take as long as 20–30 minutes to achieve full effect and the animal might be rousable even at peak effect. Animals can then be gently and quietly removed from the capture device. Relatively non-painful

procedures (e.g., blood sampling, collaring, weighing) can be performed without arousing the animal. Animals might be aroused with stimulation and the depth of sedation should be constantly monitored. Furbearers should be blindfolded and hobbled or tethered to prevent escape in the event of arousal. Caution should always be exercised with such animals even though they seem harmless. Using only alpha-2 adrenoceptor agonists in these situations allows the sedative to be completely antagonized with full recovery in just a few minutes (Kreeger et al. 1988, 1996; Baldwin et al. 2008).

Alpha-2 adrenoceptor agonists are not controlled substances. Detomidine, dexmedetomidine, and medetomidine are much more potent than xylazine and more selective for specific alpha-2 adrenoceptor receptors. Medetomidine is a racemic mixture composed of equal parts of two optical enantiomers: dexmedetomidine and levomedetomidine. The pharmacological effects of medetomidine are due almost exclusively to dexmedetomidine. Thus, the relative potency (mg:mg ratio) between dexmedetomidine and medetomidine is 2:1. Dexmedetomidine in wildlife does not seem to offer any substantial advantages over medetomidine (Fandos Esteruelas et al. 2017).

Ketamine

Ketamine is a dissociative anesthetic and is probably one of the most widely used drugs for wildlife because of its safety and efficacy. It is capable of anesthetizing all furbearing species (Kreeger and Seal 1986*a*, Kreeger et al. 1990*b*). Ketamine is characterized by producing a cataleptic state (a malleable rigidity of the limbs). The eyelids normally remain open during ketamine anesthesia and the eyes of animals immobilized outdoors should be protected from drying out and from ultraviolet light. Palpebral and corneal reflexes usually remain intact and should not be used to assess depth of anesthesia. When used alone, ketamine usually causes rough inductions and recoveries, and convulsions are not uncommon. Because of this, it is usually administered concurrently with tranquilizers or sedatives.

Ketamine is a racemic mixture composed of equal parts of two optical enantiomers: S-ketamine and R-ketamine. Recovery from racemic ketamine has been associated with undesirable psychomimetic effects in several species and S-ketamine may offer an advantage, but it is not commercially available except in intranasal form for human use. Ketamine is fairly effective when given orally and can be used in consumed baits to partially tranquilize animals, or can be sprayed into the mouth of caged or trapped animals, rendering them safer to handle. There is no antagonist of ketamine (Kreeger and Seal 1986*b*). Ketamine is currently a Schedule III controlled substance, but there is concern it will be elevated to the more highly regulated Schedule II due to abuse by humans.

Use ketamine-medetomidine combinations with extreme care when immobilizing dangerous species (e.g., gray wolves, wolverines [*Gulo gulo*]) because there have been multiple reports of sudden awakening from this drug combination. This usually occurs 30–45 minutes post-induction, and animals are capable of directed attack; however, if not further stimulated after arousal, they often revert to the immobilized state. If working with such species, constantly monitor depth of anesthesia by observing for increased respiration, head movement, ear twitching, or palpebral (eyelid) movement. Minimizing noise and limiting tactile stimulation are not recommended because the animal is metabolizing ketamine and is only heavily sedated by medetomidine, but is not anesthetized. Thus, the animal may seem to be unconscious when it is not. Talking normally and handling the animal actually promotes signs of earlier recovery, which can then be managed, than if such stimulation is minimized.

Tiletamine

Tiletamine is another dissociative anesthetic similar to, but about 2.5 times more potent than, ketamine. Tiletamine is unavailable as a single product and it is combined in equal proportions with the benzodiazepine agonist, zolazepam. Combining these two drugs results in fewer convulsions, good muscle relaxation, and smoother recoveries. Tiletamine-zolazepam is a Schedule III controlled substance.

Tiletamine-zolazepam is available worldwide in freeze-dried form (Telazol®, Zoletil®). Zoletil is produced with 500 mg (250 mg of each drug) per vial. The manufacturer recommends adding 4.4 ml solvent to the vial, resulting in a concentration of 100 mg/ml. However, Telazol is produced with 572 mg (286 mg of each drug) per vial. Manufacturer's instructions call for adding 5 ml sterile water to the vial, resulting in an approximate concentration of 100 mg/ml. This apparent inconsistency is because when 5 ml of solvent is added, the final volume is actually 5.7 ml due to chemical reactions. Thus, 572 mg in 5.7 ml is approximately 100 mg/ml (Amass and Drew 2006).

Tiletamine-zolazepam is extremely safe, having little effect on respiratory and cardiac function. In the normal concentration of 100 mg/ml, it can be used for most species of furbearers. However, for larger species (e.g., gray wolves), it may be desirable to increase the concentration to be able to decrease dart size or syringe volume. This can be accomplished by adding less water. Regardless of the amount of water added, the final volume will be approximately 0.6–0.7 ml more (e.g., adding 2 ml will result in about 2.6 ml). This total volume needs to be kept in mind because it will affect calculations. For example, when using Telazol, adding just 2 ml of water (2.6 ml final volume) will result in an actual drug concentration of about 220 mg/ml (572 mg/2.6 ml). As little as 1 ml solvent (1.6 ml final volume) can be added, but the solvent must be warm, and the solution kept warm, or the drug will go back out of solution.

The effectiveness of tiletamine-zolazepam can be increased by adding another drug (e.g., ketamine, medetomidine, xylazine) instead of water. This creates a fairly potent drug combination that is probably not necessary for most species of furbearers. It could be useful to aid in the release of non-target species (e.g., bears [*Ursus* spp.], deer [*Odocoileus* spp.]) from capture devices. Adding 2 ml of 100 mg/ml xylazine, for instance, will provide 572 mg tiletamine-zolazepam (for Telazol) plus 200 mg xylazine in approximately 2.6-ml solution. One vial of this mixture should immobilize most medium-sized (<100 kg [220 lb]) mammals; two vials should be effective on larger (100–250 kg [220–550 lb]) mammals. Alternatively, you can add 1 ml of 100 mg/ml ketamine plus 1 ml of 100 mg/ml xylazine, which is a good combination for bears. The same doses would apply (i.e., one vial) for <100-kg (220-lb) bear.

Etorphine

Opioid immobilizing agents are derived from two opium alkaloids (morphine and thebaine). The opioids have been used for animal immobilization since the 1960s and are the most potent drugs available for this purpose. A major advantage of opioids for wildlife immobilization is the availability of specific antagonists. Opioids are neuroleptanalgesics characterized by spontaneous movements and responsiveness to noise, touch, and other stimulation, which indicate that they are not completely unconscious, a characteristic of general anesthesia (Kreeger et al. 2010).

Etorphine was the first of the potent opioids to be used in wildlife immobilization. Its popularity diminished somewhat with the advent of even more potent opioids, such as carfentanil and thiafentanil. However, due to the recent banning of carfentanil and the severe restrictions on use of thiafentanil in the U.S., etorphine will again probably be the predominant potent opioid. Etorphine has always been popular in Africa and Europe. In laboratory testing of rats, etorphine was determined to be 1,000 times more potent than morphine (Dobbs 1968). Because of this potency, etorphine probably has little use for immobilization of furbearers, other than to aid in the removal of large non-target non-furbearing species from capture devices. Etorphine is a Schedule II controlled substance and should be handled with extreme care to minimize the potential for human exposure.

Sufentanil

Sufentanil is an opioid used primarily in human medicine, but it can be used on large species of furbearers (Kreeger and Seal 1990). The formulation (50 µg/ml) for humans is too dilute to be of practical use in wildlife because of the large volumes required. However, sufentanil can be purchased in bulk powder, which can be reconstituted in a variety of concentrations by veterinary compounders by dissolving in sterile water for injection and titrated to pH 4.0 with hydrochloric acid. Sufentanil is >4,500 times more potent than morphine, but has an extremely high safety index, making it the safest potent opioid in case of accidental exposure to humans (Niemegeers et al. 1976). Still, it should be used judiciously on furbearers because of the potential for severe respiratory depression. Sufentanil is a Schedule II controlled substance.

Butorphanol

Butorphanol is a mixed agonist-antagonist opioid with a potency 3.5–7.0 times that of morphine. Higher doses (>0.5 mg/kg) of butorphanol may result in no effect, as antagonistic properties tend to dominate. Alone, butorphanol provides only apathetic sedation (Kreeger et al. 1989). There is currently a resurgence in the use of butorphanol when it is combined with azaperone and medetomidine (BAM). In Canada and the U.S., it is a Schedule IV controlled substance. Advantages of BAM include smooth induction, reversibility, and fewer record-keeping requirements compared to Schedule II controlled substances (e.g., etorphine). Disadvantages include prolonged induction time (often >10 min), respiratory depression, and sudden and unexpected arousal. BAM has been used to immobilize some North American furbearers (e.g., red foxes [*Vulpes vulpes*]) and its use will undoubtedly increase in the future (Kreeger et al. 2023).

Nalbuphine

Nalbuphine is a semi-synthetic opioid agonist-antagonist that is 10 times more potent than butorphanol. It is chemically related to the opioid antagonist naloxone, and the opioid agonist oxymorphone. Nalbuphine has been shown to be a viable alternative to the more potent opioids when mixed with azaperone and medetomidine (NAM). There is one report on the use of NAM in North American beaver (*Castor canadensis*), and it has been used with somewhat limited success on gray foxes (*Urocyon cinereoargenteus*; T. Hiller, Wildlife Ecology Institute, personal communication), but its use on other mammals will probably increase (Roug et al. 2019). It is not a controlled substance in the U.S.

Alfaxalone

Alfaxalone is a neuroactive steroid with anesthetic properties. Alfaxalone was initially marketed combined with another drug. It was removed from the market due to adverse effects of the solvent. Subsequently, alfaxalone was reintroduced as a single agent utilizing a different solvent. Although usually administered IV as a preanesthetic, it can be given intramuscularly (IM). There are no reports of its use on North American furbearers, but it should be applicable because it is approved for use in domestic cats and dogs and has been used on domestic ferrets (Milloway et al. 2019) as well as small mammals elsewhere in the world (Sauvé et al. 2021). Alfaxalone combined with azaperone and medetomidine (AAM) has been effective on captive and free-ranging deer (Pon et al. 2016, Mathieu et al. 2017). Alfaxalone has an extremely brief shelf life once the vial seal has been broken. In the U.S., a vial can be kept ≤6 hours after its first use. However, in Australia and New Zealand, a used vial can be kept refrigerated for ≥7 days. It is a Schedule IV controlled substance in the U.S.

Propofol

Propofol is an injectable anesthetic chemically unrelated to other IV anesthetics. The compound comes as a 1% (10 mg/ml) emulsion in oil. It has a milky appearance, and it is easily contaminated with bacteria and mold unless a strict sterile withdrawal technique is used. When given IV, propofol produces general anesthesia of short duration (Weaver and Raptopoulos 1990). Following induction, respiration is often depressed, sometimes to the point of apnea, but this effect lasts only about 30 seconds. Due to its rapid elimination, recovery from propofol can lead to disorientation and paddling of the limbs. The addition of diazepam (0.4 mg/kg) is often helpful in providing muscle relaxation and smoothing recovery. Propofol has had some limited use in wild ruminants and camelids (Jalanka and Teräväinen 1992), but volumes required can be prodigious in large animals. There are currently no reports of its use in furbearing species, but it could be useful in some circumstances where the animal can be restrained, and the drug administered IV. Propofol is currently not a scheduled substance in the U.S., but may become a Schedule IV controlled substance in the future.

Inhalation Anesthetics

Comprehensive instructions on inhalation (gas) anesthesia is beyond the scope of this chapter. A brief, but thorough, discussion of gas anesthesia can be found in Kreeger et al. (2023); in- depth coverage can be found in Muir and Hubbel (2013) and Grimm et al. (2015). Inexperienced users should not attempt to anesthetize animals without hands-on instruction from a veterinarian or an experienced veterinary technician. In the simplest of terms, gas anesthesia is the delivery of vaporized drugs that are breathed directly into the lungs, taken up by the blood, and delivered to the brain, resulting in general anesthesia. Elimination of the drug is mostly by a reversal of this same route.

Gas anesthesia can be used effectively on small species of furbearers (e.g., black-footed ferrets [*Mustela nigripes*], muskrats [*Ondatra zibethicus*]; Fig. 17.1) that can be placed into an induction chamber. Occasionally, a pre-anesthetic sedative may be used ahead of time to allow the animal to relax prior to full sedation with gas and help decrease dysphoria during recovery. Major advantages of gas anesthesia are: 1) fairly rapid induction, 2) ability to quickly alter the depth of anesthesia, and 3) relatively quick recovery. The preferred gas anesthetics are isoflurane and sevoflurane because of their more rapid induction and recovery times compared to other gases (Kreeger et al. 1998). Both gases require a precision vaporizer. Compressed medical-grade oxygen is used to vaporize the anesthetic and deliver it to the lungs. The anesthetic is picked up in the bloodstream and delivered to the brain. Conversely, shutting off the anesthetic vaporizer and delivering only oxygen results in fairly rapid recovery.

Unlike injectable anesthetics, where the animal is usually quite stable a few minutes after induction, animals on gas anesthesia require continual monitoring. The biggest mistake that novices make with gas anesthesia is to become distracted with other tasks and ignore the depth of anesthesia. In a very short period of time, animals can go from an acceptable level of anesthesia to respiratory arrest and death. When using gas anesthesia, a person dedicated to monitoring the patient is imperative. There are numerous manufacturers of complete gas-delivery systems, components, and induction chambers. New systems can be quite expensive, but used vaporizers and components can be acquired.

Fig. 17.1. A gas-anesthesia machine that uses isoflurane carried by oxygen to anesthetize a muskrat (*Ondatra zibethicus*). Image courtesy of A. Ahlers.

Antagonistic Drugs

An antagonistic drug is a chemical substance that binds to and blocks the activation of certain receptors on cells, preventing a biological response. Some of the more notable pharmacological developments relative to chemical immobilization of wildlife have been specific, long-lasting opioid and alpha-2 adrenoceptor antagonists. The ability to antagonize anesthesia and return the animal more quickly to physiological normalcy offers many advantages including: 1) alleviation of problems associated with prolonged recumbency, such as nerve and muscle damage, bloat, and hypothermia; 2) reduced probability of injury or death after recovery due to accident or predation because there is no residual impairment (e.g., sedation, ataxia) from the immobilizing drugs; 3) decreased probability of social rejection or interspecific strife due to quicker return to parent or group; and 4) decreased personnel and equipment time dedicated to monitoring the recovery process.

In general, opioid and alpha-2 adrenoceptor antagonists are safe, causing adverse effects only at higher doses. Remember that antagonists act on the animal and not on the agonist. Thus, it does not necessarily follow that the more potent the agonist, the higher the amount of antagonist needed. Increasing the dose of an antagonist usually does not decrease recovery times (Kreeger et al. 1987), but higher doses could prolong antagonism by maintaining serum concentrations at higher levels. When given a choice, select an antagonist that is the most specific for the receptors affected and has the longest biological life in the animal.

Intravascular injection of antagonists provides the most rapid recovery time (1–2 min), although such quick recoveries may be less smooth compared to other routes of administration. A slower recovery time (5–10 min) occurs with IM injection. Spraying antagonists intranasally can also achieve reversals, albeit somewhat slower than IM (Shury et al. 2010). A common practice was to give equal doses of the antagonist both IV and IM or subcutaneously (SC), or IM and SC. The IM or SC dose theoretically provides a slower release and thus a longer period for the antagonist to prevent recycling of the agonist; however, research on domestic goats does not support this hypothesis (Mutlow et al. 2004). Unless there is a medical emergency (e.g., apnea, choking, possibility of drowning) where the animal needs to recover quickly, there is probably no reason to administer antagonists IV. Antagonists given IM provide for a more gradual, controlled recovery and are simply easier to administer.

Yohimbine and Tolazoline

The 1980s included an explosion of scientific reports when yohimbine, a long-known plant alkaloid, and tolazoline were rediscovered as antagonists to xylazine used primarily in the chemical immobilization of carnivores and ungulates (Kreeger et al. 2023). Neither yohimbine nor tolazoline are specific alpha-2 adrenoceptor antagonists. In addition to adrenoceptor activity, yohimbine and tolazoline affect several other receptor types (e.g., cholinergic, dopaminergic, serotonergic). Because of this broad activity, these agents may cause undesirable side effects. For unknown reasons, tolazoline seems to be more effective than yohimbine in some ungulate species. Yohimbine and tolazoline should be used to antagonize only xylazine and not medetomidine or dexmedetomidine. Yohimbine and tolazoline are not controlled substances. Commercially prepared tolazoline is currently unavailable in Canada and the U.S., but possibly available through veterinary compounders.

Early on, many investigators claimed that yohimbine could antagonize ketamine-xylazine anesthesia. However, yohimbine antagonizes only the xylazine component of this combination (Kreeger and Seal 1986*b*). Yohimbine should not be administered in animals anesthetized with xylazine-ketamine combinations until ≥30 minutes have elapsed since the last injection of ketamine. This is to allow further metabolism of the ketamine component of the combination. If yohimbine (or any alpha-2 adrenoceptor antagonist) is given when ketamine serum concentrations are still high, the xylazine component will be antagonized, resulting in an anesthetic recovery from what is essentially pure ketamine. Such recoveries are characterized by uncontrolled, often violent, body movements, severe hyperthermia, or both, which can cause injury or death to the animal.

Atipamezole

Atipamezole is the most potent and selective alpha-2 adrenoceptor antagonist currently known. Atipamezole effectively antagonizes the pharmacological effects of all the alpha-2 adrenoceptor agonists, and it is the preferred antagonist for all alpha-2 adrenoceptor sedatives. This drug was developed as a specific antagonist to medetomidine, and it is generally administered at a dose of 5 mg/mg of the total dose of medetomidine. For dexmedetomidine, a dose ratio of 10:1 (mg:mg) is used. Atipamezole can also be used to antagonize the effects of other alpha-2 adrenoceptor agonists: 4–5 mg/mg of detomidine and 1 mg/10 mg of xylazine (Kreeger et al. 2023). The same ratios are used also when the apha-2 adrenoceptor agent is combined with another drug, such as ketamine. The recommended route of administration for atipamezole is IM. Atipamezole is not a controlled substance.

Medetomidine is much more potent than xylazine. This potency greatly reduces the amount of ketamine required for immobilization, often by 50% or more. Animals begin to metabolize ketamine almost from the time it is administered. Thus, over time, much less active ketamine is in the animal's system compared to the amount of ketamine that would be given in a comparable ketamine-xylazine immobilization. When atipamezole is given to antagonize the medetomidine, there is much less ketamine to maintain immobilization. Because of this, recoveries are usually much faster and more complete compared to immobilizations of ketamine-xylazine with yohimbine antagonism.

However, the downside to this better recovery is that, as ketamine is metabolized, the animal is increasingly under the influence of medetomidine. Although medetomidine is a potent sedative, the animal might overcome its effects if appropriately stimulated. This can lead to a critical situation if ketamine-medetomidine is used on dangerous animals. When using ketamine-medetomidine, you should constantly monitor the depth of immobilization by checking for ear twitches, jaw tone, and palpebral reflex. Loud noises can be sufficient stimulation to arouse an animals from what is primarily a medetomidine-induced immobilization. Administer more ketamine if more time is needed, otherwise you should quickly finish processing the animal.

Flumazenil and Sarmazenil

Flumazenil and sarmazenil are potent and specific benzodiazepine antagonists that can be used for reversal of the central sedative actions of benzodiazepine agonists. Benzodiazepine antagonists may be useful in felids (but not in canids) anesthetized with tiletamine-zolazepam because the elimination time of tiletamine in felids is shorter than that of zolazepam (vice versa in canids). The disadvantage of both antagonists is that resedation tends to occur because they have a shorter half-life than most agonists. These are not controlled substances.

Diprenorphine

Diprenorphine was developed years ago as an antagonist to etorphine and should be restricted to that use. Diprenorphine acts antagonistically at the mu receptor while exhibiting agonistic properties at the 2 remaining opiate receptor sites. Thus, at higher doses, they cause agonistic effects, such as respiratory depression. Pure antagonists (e.g., naloxone, naltrexone) exhibit only antagonistic properties at all 3 opioid receptors. Diprenorphine is a Schedule II controlled substance.

Naltrexone

Naltrexone has an antagonistic activity 2–9 times greater than that of naloxone, an opioid antagonist widely used in human medicine (Bryson 1989). Besides being more potent, naltrexone has a much longer duration of action than naloxone in most wildlife species, and therein lies its advantage for use in chemical immobilization of wildlife. Naltrexone can antagonize etorphine at 20 mg of naltrexone for every 1 mg of etorphine administered, and a fixed dose of 25 mg of naltrexone is given to antagonize butorphanol in BAM (Kreeger et al. 2023). Naltrexone is superior to diprenorphine because it has a high therapeutic index, and it is the antagonist of choice for accidental exposure to humans. Naltrexone is not a controlled substance.

Doxapram

Doxapram is a CNS stimulant with a long history of use in veterinary medicine. Due to its CNS stimulation, doxapram has been used as an arousal agent in animals sedated with xylazine. However, doxapram has no specific antagonistic properties and it is used only to counteract respiratory depression during anesthesia, especially in field-emergency situations. After IV administration (0.5–2.0 mg/kg), doxapram effectively increases the rate and depth of respiration, but the duration of action is short-lived.

Analgesic Drugs

Analgesics diminish sensation to pain without the loss of consciousness. Analgesics should be provided to any animal that may perceive pain upon recovery from immobilization. Such pain may be due to a severe wound that required treatment, tooth extraction for aging purposes, or any surgical incision (e.g., transmitter implant). Although there is little known about the efficacy of analgesics in wildlife, a great deal is known about their efficacy in domestic and zoo animals and there is no reason to think that the pathophysiology of pain is different in free-ranging animals. Just because a captured animal survives the capture event, does not mean that it is functioning or behaving normally upon release.

Reasons to use analgesics include: 1) the ethical responsibility to the animal, 2) its use does no harm to the animal even if analgesic efficacy is dubious, and if nothing else, 3) an Institutional Animal Care and Use Committee (IACUC) will require it, if you are subject to approval from an IACUC. Many immobilizing drugs (e.g., alpha-2 adrenoceptor agonists, cyclohexanes, opioids) have analgesic properties, but these properties are negated when antagonists are given or when these drugs themselves are metabolized. A thorough coverage of pain and pain management is beyond the scope of this chapter, but excellent information can be found in Grimm et al. (2015) for in-depth coverage of analgesic drugs and doses.

Opioids

Opioids (e.g., buprenorphine, butophanol, hydromorphone, morphine) for pain management should be used judiciously in free-ranging wildlife due to their potential for prolonged sedation. Sedation could be problematic for prey species or for predators (e.g., gray wolves) potentially experiencing intraspecific aggression.

Nonsteroidal Anti-inflammatory Drugs

Nonsteroidal anti-inflammatory drugs (NSAIDS) are probably the drugs of choice for free-ranging wildlife. Meloxicam, carprofen, and ketoprofen are widely used in free-ranging and zoo wildlife. Ibuprofen should not be used in carnivores due to its side effects (e.g., gastrointestinal ulceration, liver and kidney effects).

EQUIPMENT

Chemical Immobilization Equipment

Most species of furbearers will probably be initially captured in traps, followed by drugs injected by hand or using a pole syringe (i.e., jab stick). Furbearers captured in foothold traps or foot-snares can be restrained by a trained person wearing appropriate handling gloves, or restrained with a catch pole, fishing net, or forked stick (Fig. 17.2). Larger, more dangerous species of furbearers can be darted using blow pipes, dart pistols, or dart rifles. Free-ranging wolves are often darted from helicopters using dart rifles (see Boyd et al. 2023 [Chapter 32]), but smaller furbearers, such as Canada lynx (*Lynx canadensis*) and wolverines, have also been darted from the air. A more thorough discussion of immobilizing equipment can be found in Kreeger and Arnemo (2018).

Syringes

Handheld syringes and needles are the basis for any drug-delivery system. Not only are they used to administer drugs directly to restrained animals, but they are also used for measuring and loading immobilization drugs into other delivery devices, such as darts. Syringes and needles are also required for collecting blood samples and administering antibiotics and other drugs. Most syringes and all needles are sterilized and disposable, and are intended to be used once and safely discarded, such as using a sharps container. In some cases, however, syringes may be used more than once if they are used to withdraw the same drugs needed for filling darts. Such syringes should be permanently labeled to identify the drug for which the syringe is dedicated. Needles should be used to withdraw or administer only one type of drug, not be used on more than one individual animal, and should not be reused for any reason. The basic philosophy is to avoid cross-contamination of either drugs or animal fluids. Needle sizes are

Fig. 17.2. Gray wolves (*Canis lupus*) and other furbearing species may be restrained with a forked stick and fishing net. Such restrained wolves can be safely hand-injected for immobilization.

defined by outside diameter and length. In the U.S., diameters are measured by gauge, where the smaller the gauge, the larger the outside (and inside) diameter (e.g., an 18-gauge needle has a larger diameter than a 23-gauge needle). The rest of the world uses the metric system to measure needles. If needed, a gauge-metric conversion chart for needles is available (Sigmaaldrich 2019).

When working at different altitudes, insert a needle into the air space at the top of the drug vial to equalize air pressure. For IV injections or for collecting blood samples, use smaller needles as appropriate for the size of the animal. Common veins from which to draw blood samples are the cephalic and jugular (Fig. 17.3). Other veins accessible in larger animals are the femoral, running along the inside of the thigh, and the saphenous, located along the outside of the hock. Keep the protective cap on the needle until just before injecting the drug, as accidental needle jabs are the number one cause of accidental human exposure (Petrini et al. 1993).

Pole Syringes

Pole syringes are very useful for administering drugs to trapped animals or safely administering additional drugs to animals not completely immobilized, but approachable (Fig. 17.4). Pole syringes are usually limited to administering <10 ml of drug because the animal will usually not hold still long enough to be given larger volumes. Manufactured pole syringes are generally superior to homemade ones; however, most pole syringes are too large in diameter to fit between the mesh of cage traps to inject captured furbearers. Smaller-diameter pole syringes can be fabricated using 1–3 ml syringes. Use large-gauge needles on large (>30 kg [66 lb]) animals and smaller-gauge needles on smaller animals. Inject the contents IM rapidly and firmly and withdraw before the animal can bite the pole. The muscle masses of the hindquarters are the preferred location for injection, but the shoulder muscles of larger animals can also be used.

Blow Pipes

Blow pipes, or blow guns, are useful devices for delivering small volumes of drugs at short-to-medium distances. They operate by propelling a dart through a pipe or tube either by rapid expulsion of the operator's breath, by compressed atmospheric air, or by compressed CO_2 via disposable cylinders. Blow pipes usually consist of 1- or 2-piece tubes measuring ≤2 m (6.6 ft). Most blow pipes propel 10-mm-diameter darts with a maximum capacity of 3 ml of drugs. The effective range of blow pipes is limited to about 20 m (66 ft).

The use of blow pipes is quiet, and darts propelled by blow pipes usually cause little trauma to the animal because the dart neither strikes the animal with much velocity nor does the method of dart operation cause injury; animals as small as 3 kg (6.6 lb) can be safely treated. Blow pipes are used primarily on captive or restrained animals, but they can also be used effectively on free-ranging animals under the right circumstances, such as animals that have been trapped or that have been treed through the use of trained dogs.

Blow pipes that use compressed air or CO_2 have valves to allow for their pressures to be adjusted for different distances. The addition of a holographic sight to a blow pipe greatly increases the ability of the user to accurately place the darts (Fig. 17.5). Such powered blow pipes propel the same type of lightweight darts (10–11-mm diameter; 1–3 ml drug volume) as conventional blow pipes, and they are preferred for delivering larger volumes of drugs at longer distances. Because of their portability and low-impact delivery of darts, adjustable CO_2- powered blow pipes should be given serious consideration for the chemical immobilization of furbearers. However, blow pipes can be relatively expensive. Use of blow pipes in some U.S. states, and in some countries (e.g., Canada, Norway) is prohibited without first obtaining a special permit from the proper authorities.

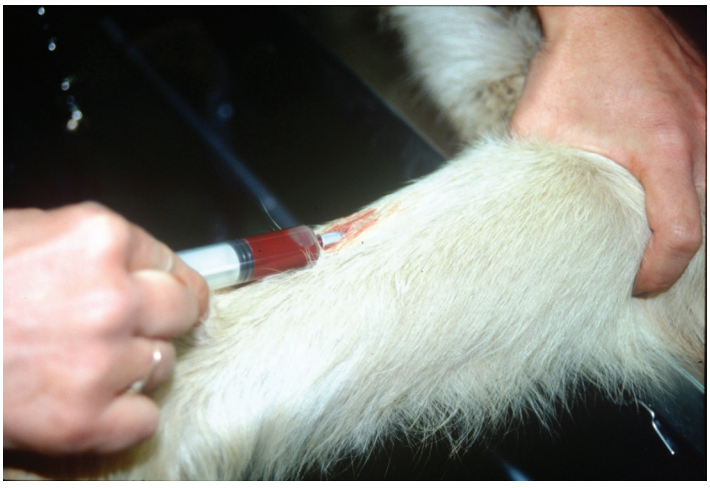

Fig. 17.3. Cephalic veins can be easily accessed in most carnivores for the purpose of collecting blood samples or administering drugs intravenously.

Fig. 17.4. Expandable pole syringes are useful tools for injecting drugs into trapped animals.

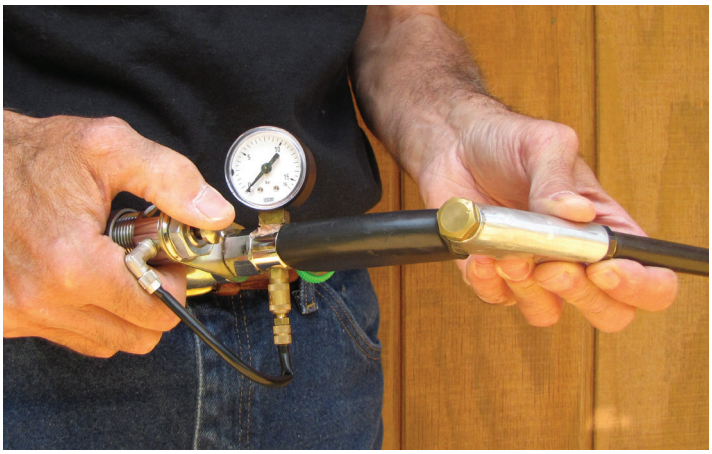

Fig. 17.5. Adjustable, CO_2-powered blow pipes can be used to dart all species of furbearers weighing >3 kg (6.6 lb); holographic sights greatly increase the ability of the user to accurately place the darts.

Dart Guns

Dart guns propel darts by either the gas generated from a .22-caliber blank cartridge, compressed CO_2, or compressed atmospheric air. Effective ranges are ≤75 m (246 ft). Dart volumes can range from 0.5 to 25.0 ml. Some manufacturers of adjustable CO_2 rifles provide 2 different sized barrels (11 and 13 mm) that can shoot a variety of lightweight plastic darts at close ranges as well larger darts at longer ranges. Such rifles, if you can afford them, can safely be used for almost all furbearers weighing ≥3 kg (6.6 lb; Fig. 17.6).

Darts

Darts can be thought of as flying syringes, consisting essentially of a needle, body, plunger, and tailpiece. Darts discharge their contents via expanding gas from an explosive powder charge, compressed air, butane, or chemical reaction (Fig. 17.7). Plastic darts using compressed air to discharge drugs are probably the preferred dart to use on small- and medium-sized species of furbearers because of the low weight and atraumatic discharge associated with these darts. Darts using explosive charges can expel their contents in <0.001 second, and thus require large-gauge needles to allow for the rapid expulsion of liquid, and are generally restricted to use on larger species of furbearers. Pneu-Dart (Williamsport, Pennsylvania, USA), however, manufactures explosive-charge Slo-Inject™ darts utilizing side ports and other technology to decrease the velocity of drug expulsion by 33%.

Needles are designed to either expel contents from the standard front opening (end port) or through a side port with the front opening usually occluded. Either wire barbs or metal collars are used to securely retain the dart in the animal. Barbs probably should be used in most circumstances because if the dart contents are under high pressure, non-barbed needles can quickly propel out of the animal due to the expulsion of the liquid, and therefore not inject any or all of the substance. Darts with VHF transmitters are also available, which helps with locating a darted animal, although these heavier darts may be better suited for larger wildlife species.

Fig. 17.6. Examples of dart guns, including from top left: Cap-Chur Short Range Projector (CO_2 powered), Pneu-Dart X-2 (CO_2 powered), Dan-Inject (CO_2 powered), Pneu-Dart model 389 (.22 caliber-blank powered), and Pneu-Dart X-Caliber (CO_2 powered).

Fig. 17.7. Types of darts, including (top to bottom): Cap-Chur (powder charged), Dan-Inject (compressed air), Pneu-Dart (powder charged with Slo-Inject®).

Support Equipment

Support equipment is important for monitoring immobilized wildlife. This includes concerns associated with thermoregulation and respiration.

Thermometers

Many immobilizing agents disrupt the thermoregulatory capability of an animal. Additionally, the physical exertion of being chased or restrained prior to immobilization often results in elevated body temperatures of the animal. Either hyperthermia or hypothermia can be fatal for an animal. Thus, monitoring rectal temperatures is important. The glass mercury thermometer has pretty much been replaced by inexpensive, electronic digital thermometers. Batteries have a habit of expiring at the most inopportune moments and thermometers tend to disappear in the field, so have several of each on hand. Some types have a long, flexible temperature probe, which allows greater probe insertion for large animals (essential to obtain accurate temperatures) or for protection of the primary electronics by placing it away from the animal.

Pulse Oximeters

Pulse oximeters are electronic devices that measure the percent oxygen saturation of hemoglobin in the blood (SpO_2). Pulse oximeters provide information on the respiratory function of the animal, which can be useful because many immobilizing drugs depress respiration. Oximeters use a clip that can be attached to the tongue or other thin, non-pigmented tissue or use of a rectal probe (Fig. 17.8). The measurement of SpO_2 is determined by passing two wavelengths of light, one red and one infrared, through body tissue to a photodetector. The oximeter processes these signals, separating the time-invariant parameters (tissue thickness, skin color, light intensity, venous blood) from the time-variant parameters (arterial blood, SpO_2). Because oxygen-saturated blood predictably absorbs less red light than oxygen-depleted blood, oxygen saturation (as well as the pulse) can be calculated.

In veterinary medicine, hypoxemia is defined when the SpO_2 is <95%, and supplemental oxygen should be given to ensure adequate oxygenation of tissue (West et al. 2014). However, it is not uncommon for animals anesthetized with opioids or alpha-2 adrenoceptor agonists to have SpO_2 values <95%. These lower SpO_2 values are usually due to temporary respiratory depression upon induction as well as acute vasoconstriction in the presence of alpha-2 adrenoceptor agonists, which should rebound as the animal resumes normal breathing, blood pressure, and heart rate. Absolute SpO_2 values are not as important as the trend of SpO_2 values. That is, if the SpO_2 steadily decreases, it can be presumed that the animal is in some sort of respiratory crisis and immediate action is necessary.

Supplemental Oxygen

If at all possible, have a source of supplemental oxygen available. Respiratory depression or arrest is the most common medical emergency encountered with chemical immobilization. All opioids and alpha-2 adrenoceptor agonists can cause some level of respiratory depression. A variety of medical oxygen tanks and delivery systems are available from medical-supply stores. Oxygen tanks carried aboard helicopters must be approved for aircraft. Although more expensive, oxygen concentrators can address this problem, and also can be used when oxygen tanks are not available (Fahlman et al. 2012). Concentrators produce a sufficient flow of oxygen for all species of furbearers, but flow may be insufficient for large animals (Fig. 17.9). Concentrators also may not perform well at very low ambient temperatures or at very high altitudes, but this is dependent on the model.

Fig. 17.8. Pulse oximeter measuring oxygen saturation in Canada lynx (*Lynx canadensis*) during chemical immobilization. Top number is oxygen saturation (%) and bottom number is pulse rate. Image courtesy of J. Arnemo, Inland Norway University of Applied Sciences, Norway.

Fig. 17.9. Oxygen concentrator used on European brown bear (*Ursus arctos arctos*) to provide sufficient oxygen flow during chemical immobilization. Image courtesy of J. Arnemo, Inland Norway University of Applied Sciences, Norway.

CAPTURE-RELATED STRESS AND MORTALITY

There are those people who obsess about stress regarding the capture and handling of wildlife. This is probably because they view stress entirely as a negative consequence. This is unfortunate because this position ignores eustress (i.e., good stress), which can result in increased motivation and productivity that can be important for survival. Any discussion of stress should include a definition of stress, but there has been no definition that is universally accepted. There are many factors which influence any definition of stress. There is physical stress (e.g., fighting, running, struggling) and psychological stress (e.g., fear, panic, unease). Presumably, the capture of animals always has components of both.

One should also differentiate between acute stress versus chronic stress. Acute stress activates the sympathetic nervous system and adrenalin secretion in the fight-or-flight response. Acute stress is of short duration (i.e., minutes) and usually does not result in measurable pathology. Chronic stress is of much longer duration (hours, days, or even years) and often causes pathology measured by immunosuppression, infertility, and loss of fitness. Thus, for the purposes of most animal captures, stress can be defined as the internal physiological and biochemical changes that alter homeostasis when an animal is subjected to physical and psychological perturbations.

Every method of animal capture causes acute stress; chronic stress probably only occurs if an animal is held in captivity or transported for long periods of time. There is no way to avoid stressing the target animal. The physical and psychological stress for wildlife associated with capture goes without further elaboration, and there is no way to eliminate all stress from any form of animal capture (Kreeger et al. 1990*c*, White et al. 1991). Capturing the animal quickly and efficiently, and releasing it as soon as possible can at least minimize stress. This can be accomplished by: 1) using the correct amount of drug (i.e., do not underdose); 2) using the most potent drug available; 3) processing the animal quickly (i.e., be organized and efficient); and 4) using drugs capable of being antagonized, if at all possible, so that the animal can return to normalcy quickly.

Chemical immobilization is a potentially life-threatening undertaking for an animal. Although death directly due to the drug(s) is actually quite rare, death due to the process of chemical immobilization is not uncommon. Animals can die during induction by drowning, falling from heights, or suffocating (e.g., neck bent or draped over an object). Upon recovery, animals can succumb to the effects of hyperthermia or hypothermia, stress, or predation. Most of these causes of mortality can be avoided through user training, experience, careful preparation, good planning, and patience. There are estimates of mortality rates associated with chemical immobilization based on large data sets collected for some species of furbearers. Arnemo et al. (2006) estimated capture-related mortality rates of three furbearing species in Scandinavia, which included Eurasian lynx (*Lynx lynx*; 3.9%; $n = 380$), gray wolves (3.4%; $n = 89$), and wolverines (2.8%; $n = 461$).

Eventually, it will be only a matter of time before an animal dies due to chemical immobilization. If one cannot afford to have an animal die because of politics, publicity, philosophy, or economics, then do not immobilize the animal.

DRUG-INJECTION LOCATIONS ON ANIMALS

Immobilizing drugs are almost always administered IM. The usual injection locations are the large muscle masses of the proximal hindlimb and forelimb, with the former being the most commonly used. Hindlimb injections should preferably be placed towards the rear so as to avoid the sciatic nerve and femur; forelimb injections should be placed towards the front. Areas of large fat deposits should be avoided, as drug absorption from these sites is slow and unpredictable. For example, bears should be injected in the lower regions of the hindlimbs or shoulder to avoid the fat deposits around the rump. For darting efforts from helicopters, the hindlimb muscles or the back muscles running along the rear one-third of the animal are suitable targets (Fig. 17.10).

Intravascular Injection

Intravascular administration is usually reserved for antagonists. Intravascular administration of anesthetics should be done with caution because the onset of action is often quite rapid and, in some cases, respiratory depression or arrest can occur. However, IV administration of immobilants may be necessary if the animal is recovering quickly and more time is required to process it. Any drug containing propylene glycol (i.e., diazepam) should be given slowly IV because a bolus can cause cardiac arrest. Intravascular administration of antagonists could result in very rapid recovery; be sure that you have an unobstructed escape route in mind and that all hobbles, snares, and blindfolds have been removed from the animal.

Fig. 17.10. Darting via helicopter is practical for medium-to-large furbearers, such as gray wolves (*Canis lupus*; shown), Canada lynx (*Lynx canadensis*), and wolverines (*Gulo gulo*). Image courtesy of L. Gangås, Statens naturoppsyn (Norwegian Nature Inspectorate), Norway.

Intranasal Administration

Spraying drugs into the nose, particularly the alpha-2 adrenoceptor sedatives, has been shown to have a substantial calming effect on physically restrained animals. Intranasal administration is usually achieved by attaching a syringe to a catheter and inserting the catheter into the nares and extending it to the level of the eye, but be prepared for violent sneezing, which may spray the drug. Have some air in the syringe to fully expel the drug when using a long feeding tube. Intranasal drugs are easier to administer to an awake, struggling animal than IV administration and onset (60–90 sec) is much faster than IM injection.

Oral Administration

Oral administration is not often used in wildlife capture primarily because of the difficulty in predicting the dose that the animal receives. Also, drugs taken orally have variable absorption rates, resulting in prolonged and erratic induction and recovery times. Drugs can be placed in tabs that are attached to traps so that when a captured animal bites on the tabs, it ingests the drug (e.g., diazepam for coyotes [*Canis latrans*] and foxes). Drugs can also be placed in food baits, but it is particularly difficult to predict the administered dose with this approach. Gray wolves have been heavily sedated by placing concentrated tiletamine-zolazepam into bait (T. Kreeger, University of Minnesota, unpublished data). If other methods fail for administering drugs, drugs may be sprayed into the mouth of the animal (West et al. 2014).

MONITORING IMMOBILIZATION AND RECOVERY

Signs of Immobilization

Familiarity with the signs of anesthesia is essential. You can assess drug effect through changes in animal behavior, but to determine such effects, it is critical to be familiar with the target species. Know what is normal and look for the abnormal. Once the animal is down, you need to assess the depth of anesthesia. Initially, observe for spontaneous, non-repetitive movements and, if present, you can usually assume that the animal is not fully immobilized. Repetitive, stereotypical movements may occur when using opioid agents. Such animals are effectively immobilized, but can still bite. If the animal seems to be unconscious, check for ear twitch (touch inside of ear and observe for ear movement), pedal reflex (pinch toe and limb withdraws), swallowing reflex (pull tongue, release, and animal swallows), jaw tone (spread jaws and feel for resistance), palpebral reflex (touch eyelashes and animal blinks), and corneal reflex (touch cornea and animal blinks). If the animal does not exhibit the ear twitch, it is probably at an appropriate stage of anesthesia for most field procedures. The cyclohexanes often do not abolish the blinking reflexes, even when the animal is fully anesthetized.

Addressing Incomplete Immobilization

If at all possible, record the time when the drug was first injected. Allow 10–15 minutes to elapse after the first injection before giving booster doses. Exceptions to this recommendation are when using drug combinations containing an opioid agonist-antagonist (e.g., BAM, NAM) or immobilizing species with relatively high levels of body fat, such as bears. In these cases, extend the period to 20 minutes. However, in almost all cases, the animal will be showing signs within the first 10 minutes. If the animal is showing signs of receiving some of the drug, administer a booster dose at 50% of the original dose. If a combination of a primary immobilant (e.g., ketamine) plus a tranquilizer (e.g., medetomidine, xylazine) was used, only booster with a half dose of the immobilant and no more tranquilizer. However, it is still perfectly safe to administer half of the tranquilizer as well.

When an animal is showing some drug effect, but is not laying down, try to minimize further stress. If an animal tries to get up when approached, retreat immediately and continue observing. If no sign of drug effect is apparent after 10 minutes, assume that the animal probably received little or none of the original dose. If the drug(s) and dose(s) that originally selected were appropriate, then give the animal the same drug(s) and dose(s) again.

Animals can be kept immobilized for extended periods (several hours) with supplemental boosters of the initial immobilizing dose. When ketamine is given initially in combination with another agent, such as xylazine, usually only the ketamine needs to be given to maintain immobilization. If the animal starts to arouse (e.g., ear twitching, head movements), IM injections of ketamine can be given as many times as needed to maintain immobilization. Smaller species of furbearers (e.g., foxes) may require as little as 25 mg of ketamine, whereas larger species of furbearers (e.g., gray wolves) may require ≤100 mg of ketamine. If the animal is laying down and can be handled, but it continues to struggle and is generally difficult to handle, a low dose of a tranquilizer, such as medetomidine (e.g., 1 mg), or midazolam (0.1–0.5 mg/kg) often calms the animal enough to allow safer and easier handling.

Handling Immobilized Animals

When an animal is finally laying down and can be safely handled, there are several immediate steps that need to be taken before other actions take place. This includes actions such as radiomarking and collection of data.

Body Position

Ensure that nothing impinges on breathing and that the neck is straight, the nose is clear, etc. Furbearers may be positioned in any position, but on their side is the most common. Preferably, the head should be higher than the thorax, with the nose pointing down to avoid aspiration of fluids. Try to keep the animal on a relatively flat surface to avoid occlusion of the trachea, pressure neuropathy, or circulatory impairment.

For lengthy immobilizations, roll the animal on its other side or sternally at least once every 60 minutes. Covering the eyes protects them from harmful ultraviolet light from the sun, reduces drying, and prevents dirt and debris from entering them. Coating the eyes with a lubricant further prevents drying; however, some believe that eye ointments result in dirt and grit sticking to the eye. A saline wash (e.g., contact lens saline) can also be used. Covering the eyes also seems to further calm the animal even though it is

effectively immobilized. If plugging the animal's ears with cotton or cloth to avoid response to sounds, which can happen with animals given opioids, attach the plugs to each other, mark with a bright string or ribbon so as not to forget to remove them, or do both. Generally, furbearers do not need to be hobbled, but a limb may be secured if sudden arousal and escape is a concern (e.g., when using only alpha-adrenoceptor tranquilizers or low doses of ketamine).

Check Vital Signs

Once assured that the animal's body position will not affect breathing, check its respiratory rate (RR). Regardless of claims, there are very few scientifically measured normal resting RRs of undrugged wild animals. Experience with a given species and capture process is the best guide. Respirations can be observed (movement of the animal's abdomen or chest), felt (place your hand in front of the animal's nostrils), or heard (place your ear by the animal's nostrils; a very sensitive technique). Slowed RRs are most likely drug-induced, but they can also be caused by hypothermia. In cases of respiratory arrest or poor oxygenation, respiration can be supported mechanically or pharmacologically. Rapid RRs could indicate hyperthermia, bloat, aspiration, pulmonary edema, or shock. Use other parameters (e.g., body temperature) to differentiate the causes of a rapid RR and treat accordingly.

Use a stethoscope to listen for abnormal chest sounds, such as gurgling, which may indicate pulmonary edema. If the animal's gums (or other mucous membranes) are pinkish (as opposed to blue or gray), tissues are probably adequately oxygenated, even if the RR is 5–6/min. Depth of respiration is as important as rate. An adequate rate with a shallow depth may result in a low volume of air being moved, resulting in an increase in CO_2 concentration in the blood.

Carry a thermometer and use it continually throughout the immobilization period. In general, rectal temperatures of mammals range from 37.5 to 38.8° C (99.5–101.8° F), but normal temperatures should be established for each species. Also, rectal temperatures do not accurately reflect core body temperatures, which are usually higher. The farther the thermometer probe can be inserted, the more accurate it will reflect true core temperature. Even then, the probe should be in contact with rectal tissue and not in an air space or feces. Temperature can change with season, and not just for hibernating animals.

There are also very few instances of scientifically measured normal resting heart rates of wild animals, so let experience be the best reference. Smaller animals generally have higher heart rates than larger animals. Heart rates can be detected by: 1) using a stethoscope (usually best detected on the down side of the animal, between the fourth and sixth ribs or behind the point of the elbow); 2) feeling the heartbeat directly by compressing the chest slightly; 3) locating an arterial pulse; or 4) using a pulse oximeter or electrocardiogram. A very fast heart rate could be a function of drugs (e.g., ketamine), physiological responses (i.e., stress, excitement), hyperthermia, or shock. Use other parameters to differentiate the causes of tachycardia. An abnormally slow heart rate could be a function of drugs (e.g., xylazine), hypothermia, or metabolic disorders (e.g., hypercalcemia, hyperkalemia). Generally, if the capillary refill time (see below) is <2 seconds, adequate perfusion (delivery of blood) is assumed, and no action is required in the absence of other signs.

Recovery from Immobilization

An animal recovering from anesthesia generally should not be left unattended. Ideally, it should be monitored until it can walk in a relatively coordinated manner (i.e., respond appropriately to objects, people, or other animals), regardless of whether an antagonist was administered. At a minimum, stay with the animal until it can at least raise itself to a sternal position. An obvious exception to this is when handling dangerous animals, such as bears (but not wolves). If unable to remain with the animal throughout recovery, place it in a dry cool or warm place, depending on weather conditions (i.e., out of the sun in summer, in the sun during winter) and free from interspecific or intraspecific harassment or aggression. Note any potential hazards such as sharp rocks, ledges, and especially water, in the recovery area. Either relocate the animal or stay with it through recovery to direct it away from such hazards.

EUTHANASIA

Invariably, there will come a time when an animal must be euthanized because it has been critically injured (e.g., struck by a vehicle) or it is terminally ill. If an animal needs to be euthanized, it should be done safely and effectively with some consideration for the dignity of the animal and the sensitivities of the public. Many methods of euthanasia, such as shooting and stunning, are effective and medically acceptable, but may be considered reprehensible to the public. Chemical euthanasia is generally the preferred method because it is safe, effective, and aesthetically acceptable. Listed below are the various methods of euthanasia that are generally employed for wildlife. Other methods, such as CO_2 and inhalant anesthesia, are not listed only because they are not practical for field application. A detailed discussion of euthanasia methods is published by the American Veterinary Medical Association (2013). The following euthanasia methods are those recommended by the AVMA, but these are only recommendations and do not have any regulatory force.

Cervical Dislocation

Cervical dislocation can be used to euthanize small rodents and rabbits, and perhaps smaller furbearers such as weasels (e.g., *Mustela* spp.). For example, for mice and rats, the thumb and index finger are placed on either side of the neck at the base of the skull. With the opposite hand, the hind limbs are quickly pulled, causing separation of the cervical vertebrae from the skull. For small rabbits, the head is held in one hand and the hind limbs in the other. The animal is stretched, and the neck is hyperextended and dorsally twisted to separate the first cervical vertebra from the skull.

Decapitation

Decapitation is generally not acceptable due to animal (and public) distress.

Exsanguination

Exsanguination (bleeding to death) is acceptable only if the animal has been rendered unconscious by drugs. It is often a slow, messy, and unsightly process. Bilateral sectioning of the jugular or femoral veins can be effective, but often the blood flow slows after clotting initiates. If possible, try to severe the major arteries leading from the heart by inserting a long-bladed knife into the junction of base of the neck and shoulder and slicing inwards and downwards.

Stunning

Stunning by a sharp blow to the head with a hard object can kill most species of furbearer. The animal should be anesthetized according to AVMA guidelines and in most cases, it is assumed that the animal will be initially chemically immobilized. The disadvantage of any method of stunning is that it may not cause death, so check that the animal is dead by monitoring heart rate, respiration, or pupillary reflex.

Gunshot

Gunshot is often the most practical, if not only, means of euthanizing wild animals. Ideally, the animal is under some sort of physical or chemical control so that carefully placed shots can be made. If the animal is not controlled, heart or lung shots are preferable to head or neck shots. Although head shots are the most sure and humane method, sometimes the head must be preserved for disease diagnoses (e.g., rabies). In these cases, the neck is the next preferred site.

Chemicals

Several euthanasia products are formulated to include a barbituric acid derivative (usually sodium pentobarbital) with added local anesthetic agents. These drugs are U.S. DEA Schedule III controlled substances. Barbiturates are generally the preferred method to euthanize domestic animals and they are acceptable for almost all species and sizes of animals. Intravascular injection is the preferred route, although intraperitoneal (IP) and intrathoracic injections can be given to small animals.

Neuromuscular blocking drugs, such as succinylcholine, may be used in anesthetized or otherwise unconscious animals. Death will be by suffocation due to muscle paralysis, thus always use an overdose for the particular species. Injections can be given IM.

Another method of euthanasia that is available to anyone is IV injection of potassium chloride (KCl). Increasing the concentration of circulating potassium (hyperkalemia) in the blood directly influences electrical activity of the heart resulting in cardiotoxicity and arrest. KCl can be inexpensively obtained from chemical suppliers, and is also available in grocery stores as light salt, which is a substitute for sodium chloride. A saturated solution can be made by adding about 300 mg KCl/ml of solvent (sterile water, physiological saline, distilled water, or even tap water). Shake vigorously and immediately draw into a syringe, as the KCl will settle out quickly of this saturated solution. This solution must be given IV (or intracardiac) quickly (slow, drawn-out administration will not be effective). Administer at a dose of 100–200 mg KCl/kg body weight. The animal should be anesthetized before KCl is administered. Cardiac arrest is quite rapid (<30 sec) and should be verified by listening for heartbeat or feeling for a pulse. Animals euthanized with KCl will often have clonic muscle spasms (arching of neck, twitching) and sometimes vocalization for a few minutes following administration.

Carcass Disposal

In the U.S., federal law mandates that animals euthanized with barbiturate solutions must be cremated or deeply buried. This is to prevent pass-along toxicity to scavengers. Barbiturate toxicity, sufficient to kill a dog, has been reported to remain in a carcass for ≤2 years after euthanasia. Animals that have received only succinycholine or KCl can safely be left in the field with no harm to scavengers.

MEDICAL TREATMENT OF FURBEARERS

This section is not intended to be a comprehensive course on veterinary emergency medicine, but is intended to address the most common medical emergencies encountered in the chemical immobilization of furbearers. The list of possible complications is lengthy, but the majority of problems are concerned with respiration and body temperature. The information provided here also includes the assumption that captures are conducted in the field where monitoring and emergency equipment might be minimal. Thus, the ability to assess problems will be limited to what can be seen, heard, or felt. The order below is loosely based on the probability of occurrence as well as necessity for immediate action.

Respiratory Depression or Arrest

Respiratory depression or arrest is probably the most common complication encountered in immobilization of wild animals. The best advice concerning respiratory arrest is to not panic. The animal probably has ≤5 minutes before irreversible, hypoxic brain damage occurs. This is really a very long time in which to take corrective action. Panic serves only to confuse your thinking and diffuse corrective efforts, both of which cost the animal time.

The main causes of respiratory depression are: 1) drug-induced depression of the respiratory center, 2) airway obstruction, and 3) pressure on the diaphragm. Signs of respiratory difficulties are: 1) few, shallow, or no respirations; 2) cyanosis (gums blue or gray); 3) noisy breathing, wheezing, rattling; and 4) oxygen saturation trend is continually downwards.

Treatment for respiratory depression includes: 1) ceasing all further administration of immobilizing drugs; 2) establishing patent (open) airway; 3) insuring that neck is straight, tongue pulled out, trachea clear of vomitus, foreign objects, etc.; 4) positioning animal correctly (furbearers can be sternal or lateral, ruminants should be sternal); 5) beginning manual chest compression by laying the animal on its side and pushing down firmly on the chest (15–20 compressions/min); 6) administering oxygen by passing a plastic tube of appropriate size into the nasal cavity, stopping the tip at the level of the eyes; 6) administering

doxapram (1–2 mg IV) if artificial resuscitation did not cause the animal to start breathing on its own; and 7) trying acupuncture by inserting a needle into the upper lip just between and below the nares (Fig. 17.11). If just the act of inserting the needle does not cause the animal to take a breath, try twirling the needle or moving the needle in and out. If neither of these actions work, relocate the needle and try again.

If artificial resuscitation or doxapram did not cause the animal to start breathing on its own, the only recourse is to antagonize any drugs that have an antagonist, even though it means that the animal might be released. If you cannot locate a vein within 30 seconds, split the dose and give the antagonist in two different locations in the shoulder or hip muscles.

Hyperthermia

Hyperthermia is elevated body temperature due to failed thermoregulation that occurs when a body produces or absorbs more heat than it dissipates. Severe hyperthermia (generally >41° C [106° F]) is a medical emergency and the animal must be cooled immediately. However, many people intervene when temperatures reach 40° C [104° F]). There is no reason not to intervene at this temperature, and such intervention may be personal preference. Obtaining a rectal temperature should be one of the first steps taken as soon as the animal can be safely handled. Monitor the temperature throughout the immobilization period.

Hyperthermia is caused by: 1) metabolic heat generated by physical exertion (e.g., struggling when captured), 2) heat absorption from environment, 3) confinement in poorly ventilated space, 4) drug-induced alteration of thermoregulatory centers, or 5) bacterial or viral infection. The signs of hyperthermia include: 1) elevated rectal temperature (>2° C [3.6° F] above normal); 2) extremities (ears, feet) very warm; 3) rapid, shallow breathing; 4) rapid heart rate, irregular pulse; and 5) coma, death.

To treat hyperthermia, first move the animal out of direct sunlight, if possible. Whole-body immersion in water is probably the most rapid means of decreasing body temperature (Fig. 17.12). Other treatment methods include: 1) spraying the entire animal with water, particularly the groin and belly; 2) packing ice or bags containing cold water on groin or head; 3) dousing the groin area with isopropyl alcohol (rapid evaporation cools quicker); 4) administering cold-water enema; or 5) administering cold lactated Ringer's solution IV or IP. Again, if all else fails, antagonize any drugs capable of antagonism.

Hypothermia

Hypothermia is a decrease in body temperature, which happens when a body dissipates more heat than it absorbs. It can be caused by: 1) drugs that decrease metabolism, endogenous heat production, or both; 2) cold ambient temperatures (<−18° C [0° F]); 3) loss of insulation (e.g., wet, soaked fur); 4) malnourishment (decreased fat); 5) recumbency in one position for too long (compresses downside fur); and 6) inadequate circulation caused by capture device. Signs of hypothermia include: 1) decreased rectal temperature (>2° C [3.6° F] below normal), 2) shivering, 3) decreased heart rate, 4) decreased blood pressure (pulse difficult to feel), 5) extremities cold to touch, and 6) extremities firm (frostbite). The only

Fig. 17.11. Respiration can be stimulated by inserting a needle in the middle of the upper lip just below the nostrils. Upon insertion, the animal should take a breath; further respirations can be stimulated by moving or twirling the needle.

Fig. 17.12. The fastest way to lower elevated body temperatures (i.e., hyperthermia) is to immerse as much of the animal as possible in water, such as shown for this wolverine (*Gulo gulo*). Image courtesy of J. Arnemo, Inland Norway University of Applied Sciences, Norway.

treatment for hypothermia or frostbite is warming the animal or affected part. Animals can be warmed by using containers of warm water, blankets, foam pads (place under animal), hand warmers, body heat (put small animal inside of your coat, or in vehicle), and electric heat pads or lights.

Regardless of the method(s) used, expect a slow recovery back to temperatures suitable for release of the animal. Antagonism of immobilizing drugs is not recommended for hypothermia cases. This is because recovery is invariably slow and if you release the animal with depressed temperature, it may walk away seemingly normal, but only to succumb to hypothermia later because it was unable to produce enough endogenous heat to re-warm itself. Boosters of immobilizing drugs might have to be given to keep the animal unconscious until warmed. However, because the animal's metabolism is decreased by hypothermia, the drug effect is usually prolonged, and recovery will be slow anyway. Hypothermia is not as common of a problem as hyperthermia, and is unfortunately often overlooked in the field.

Shock

Shock is a clinical syndrome characterized by ineffective blood perfusion of tissues resulting in cellular hypoxia. Shock can happen to furbearers which have undergone stressful or strenuous capture or handling. Shock can be caused by: 1) prolonged physical exertion; 2) prolonged physiological or psychological stress, or both; or 3) severe blood loss. Signs of shock include: 1) rapid breathing, 2) rapid heart rate, 3) low blood pressure (slow capillary refill), 4) muscle weakness, or 5) depressed sensorium (diagnosis often masked by drugs).

Treatment of shock is often unrewarding. Ideally, one would administer 10 ml/kg of IV physiologic fluids (e.g., 0.9% NaCl solution) or 20 ml/kg under the skin for physiologic support. Furbearers that have been trapped, allowed to recover, and then confined for transport can be extremely stressed, usually characterized by uncharacteristic hyperventilation. In such cases, a possible treatment would be to administer sedatives or tranquilizers to effect. Many deaths of captured animals are attributed to stress or shock, but a definitive diagnosis often remains open. As with capture myopathy (see section), there may be little that you can do to treat shock, but rather focus on prevention.

Bloat

Bloat is caused by excess gas in the stomach caused by undigested food, and the swelling stomach compresses the diaphragm and lungs to impair respiration. Bloat is not uncommon in furbearers. However, most captures will have been completed and the capture drugs antagonized or metabolized before bloat develops to the point of causing complications.

Bloat can be caused by drugs or by incorrect body position. Signs include: 1) increase in size of abdomen, 2) labored breathing (rapid, shallow), and 3) increased salivation. If at all possible, position the animal on its sternum and hold its head up to straighten the esophagus. This alone may solve the problem. If not, a stomach tube of the appropriate size may have to be inserted through the esophagus into the stomach to release intestinal gas. Apply an appropriate lubricant to the tube and be sure that there are no sharp edges on the tube that could lacerate tissue. If you are not sure about the placement of the tube, listen to the end of it for breathing sounds, which would occur if the tube is placed into the trachea rather than the esophagus. Similarly, the animal will often cough if the tube is placed into the trachea.

Vomiting and Aspiration

Vomiting is the ejection of stomach contents through the esophagus and mouth; aspiration is the inspiratory sucking into the airways of foreign material, such as vomitus. Vomiting can occur in furbearers that have recently eaten, then been chemically immobilized. Medetomidine and xylazine in particular can cause vomiting in canids and felids. Clear the vomitus, mucus, etc., as much as possible from the mouth and pharynx. Place the animal on its sternum with its neck down and head extended, then lift the body with the head and neck remaining down to help clear vomitus.

Smaller animals may be suspended by their rear legs and shaken up and down slightly. Vomiting in and of itself may not be a problem; however, aspiration of the vomitus is problematic. Not only can the animal choke on the vomit and die, the mere aspiration of only a small amount of stomach contents can inoculate the lungs with bacteria, resulting in pneumonia. The pneumonia may not develop for days, long after the animal has been released and beyond further treatment. Thus, aspiration may result in the delayed death of the animal even though at the time of recovery it seemed perfectly healthy. Antibiotics should always be administered to an animal if aspiration is suspected. Aspiration of large amounts of vomitus has a grim prognosis for the animal and euthanasia may be considered.

Capture Myopathy

Capture myopathy (CM; also called exertional myopathy) is a complex condition affecting animals which usually have undergone a particularly stressful or strenuous capture or handling event. CM is invariably associated with severe or prolonged physical exertion, but psychological stress is suspected as an important initiator of CM. Capture myopathy occurs predominantly in ungulates, but it has also been reported in birds, canids, marsupials, primates, raccoons, and seals. Fortunately, CM is rare, but not impossible, in furbearers, particularly for trapped furbearers. Signs of CM include: 1) ataxia, weakness; 2) paralysis, inability to stand; 3) stiff, hard muscles; 4) myoglobinuria (dark, brownish urine); and 5) death.

There is essentially no treatment for CM. The only reported successful treatment of a captive coyote diagnosed with CM included dantrolene (used to treat exertional myopathy in horses), IV fluids, and steroids for several days (Ashley 2018). Signs of CM may develop within a few hours of capture or may not develop for several days. Blood samples will show severely altered serum chemical values; necropsy of the hindquarters often reveals gross or microscopic muscle degeneration.

Seizures

Seizures (convulsions) are transient disturbances of cerebral function characterized by violent, involuntary contractions of the voluntary muscles. Most seizures observed during chemical immobilization are due to the use of ketamine, including in conjunction with the alpha-2 adrenoceptor or phenothiazine tranquilizers. Usually, a few seizures do no harm to the animal, but they disrupt handling of the animal and multiple seizures can lead to hyperthermia and other complications if left untreated. Seizures accompanying ketamine immobilizations are most common during induction and recovery from anesthesia.

Signs of seizures include: 1) uncontrolled muscle spasms, and whole-body spasms; 2) rigid extension of the limbs; and 3) mouth gaping. Seizures can be effectively treated by administering diazepam (5–10 mg) IV over a 10–15-second interval to prevent cardiac arrest due to a rapid IV bolus. Midazolam can be substituted for diazepam at the same dose, but midazolam does not have to be injected slowly. Both diazepam (less ideal) and midazolam can be given IM. Repeat the dose if animal continues to seizure.

Wounds

Depending on the capture device used, furbearers may have skin and tissue lacerations. Minor wounds probably require no treatment, other than antibiotics. Small, shallow lacerations can be lavaged with a povidone-iodine, 2% chlorhexidine scrub solution, or sterile saline. Deeper, penetrating wounds can be flushed by diluting povidone-iodine with sterile saline to a 10% solution or 2% chlohexidine to a 0.05% (i.e., 1:40 dilution of the 2% solution) solution. Generally, deep, penetrating wounds should not be sutured to allow for drainage; lengthy lacerations, deep or shallow, should probably be sutured. Do not suture wounds if unfamiliar with the different types of suture materials, needles, and patterns. For more information, consult surgery textbooks or, better, obtain firsthand experience with a veterinarian. Nonetheless, the situation is somewhat simplified under field situations.

Although it is generally preferred to use non-absorbable sutures to close the outer skin layer (to be removed after healing) and absorbable sutures for all internal layers, use only absorbable sutures in the field for all closures because the animal will be released, and therefore it is very unlikely that sutures could be removed after healing. If suturing a wound is required during cold winter months, do not shave the fur around the wound because the animal will lose substantial amounts of body heat, thereby causing concern for hypothermia. Rather, saturate the wound area with a mixture of iodine and sterile gel lubricant, then just push the sticky hair coat aside while suturing.

Any animal receiving a laceration should be given antibiotics to reduce the magnitude of infection. Penicillins are the most commonly used antibiotics because they are effective against many of the skin microbes as well as formulated in repository (long-lasting) preparations. A combination of procaine penicillin G and benzathine penicillin G provides both fast, high blood concentrations (procaine) and prolonged therapeutic concentrations (benzathine; 5–7 d). Penicillins need to be kept refrigerated when not in use, which is a drawback for extended field use. Cefovecin is probably the best antibiotic because of its efficacy against many bacteria and its long duration (≤14 d) after a single dose. It is, however, quite expensive; a single dose in a gray wolf could currently cost US$100.

Cardiac Arrest

Cardiac arrest is the loss of effective cardiac function resulting in cessation of circulation. Fortunately, total arrest is extraordinarily rare during chemical immobilization of wildlife. In thousands of immobilizations, I have encountered only one event of cardiac arrest, which was with a gray wolf. Dissociative drugs (e.g., ketamine, tiletamine) are cardiac sparing, but any injectable sedation or anesthesia comes with inherent cardiac risk, albeit slight. The signs of cardiac arrest include: 1) weak or absent heart sounds or pulse; 2) poor capillary refill (>2 sec; see below); 3) cyanosis (gums are blue or gray); 4) increased respiratory rate, abnormal pattern, or apnea; 5) dilated pupils; and 6) loss of consciousness.

Probably the only treatment for cardiac arrest while in the field would be to place the animal on its side and apply pressure downward over the heart. Compress for a count of one and release for a count of one with 60–100 cycles/minute. Epinephrine (0.2 mg/kg of 1:10,000 dilution) can be very effective for cardiac arrest, but it must be given IV or IC. In the field, heart function has successfully been revived with cardiac massage and ≥1 injection of epinephrine, but few drug kits will contain epinephrine (and if they did, the drug would probably have expired before use).

Capillary refill time (CRT) is a method to assess peripheral perfusion and, by inference, cardiac function. To evaluate CRT, locate a non-pigmented (i.e., pink) area on the gums, vulva, inner eyelid, or other appropriate area. Apply pressure to this site with a finger and the compressed area will turn pale due to blockage of blood circulation. Release finger pressure and time how long it takes (by counting one-one thousand, etc.) for the bloodless area to turn pink again as blood perfusion is restored. A CRT of <2 seconds generally implies adequate blood pressure. A CRT of >2 seconds indicates low blood pressure or other circulatory dysfunction.

DRUG DOSES

Drug doses for furbearers covered in this chapter are based on published reports or personal experience (Kreeger and Arnemo 2018; Table 17.1). If a species of furbearer is not listed, use the provided doses of a closely related species as a starting point. For example, no dose is listed for Russian sable (*Martes zibellina*), but a dose is listed for American marten (*Martes americana*), so use the latter dose as a starting point. These species are similar enough such that drugs and doses for one should be safe and effective for the other.

Another option to consider if you cannot find a drug dose for a particular species of furbearer is to use an initial dose of 3.0 mg/kg ketamine plus 0.1 mg/kg medetomidine (Kreeger and Arnemo 2018). An easy way to pre-mix this dose is to start with 200 mg/ml ketamine (available from veterinary pharmacists) and 20 mg/ml medetomidine (Wildlife Pharmaceuticals, Fort Collins, Colorado, USA). For each 20-ml vial of ketamine, remove 5 ml and replace with 5 ml of medetomidine (for 10-ml vials of ketamine, remove and replace 2.5 ml). This combination will now provide 150 mg of ketamine and 5 mg of medetomidine per ml.

It is also important to note that listed drug doses are guidelines for where to start, but characteristics (e.g., age, reproductive status, overall health, level of stress) of individual animals should be considered to determine whether a lower or higher dose should be used. In general, debilitated animals should be dosed lower. Similarly, external factors such as chase times or whether an animal is already highly excited before approached, may require a higher initial drug dose for an animal.

CONCLUSIONS

This chapter was written to familiarize the reader with the essentials of chemical immobilization. Inexperienced personnel should endeavor to gain a more in-depth knowledge of drugs, equipment, and techniques through additional reading and training. Even if you have had some general experience with chemical immobilization, there is no substitute for working with someone with direct experience in capturing and handling the species of interest. This does not have to be a veterinarian; some of the most knowledgeable professionals in capture and immobilization are field biologists with years of experience with a single genus.

It is unlikely that the need for capture and chemical immobilization will dissipate in the coming years. As just one example, the incredible life-history data that can be collected from animals through the use of modern GPS transmitters will continue to require the capture and safe handling of wildlife. However, the continual abuse of immobilizing drugs by a substantial number of humans has already eliminated some drugs and threatens to eliminate some critically important drugs (e.g., ketamine). Researchers will hopefully counter these losses with development of new combinations of drugs (e.g., BAM, NAM) or perhaps entirely new drugs (albeit unlikely). The challenges associated with prevalent drug abuse by humans, terrorism, government regulations, and the increasing costs of development and approval of drugs could severely limit access to and use of drugs for chemically immobilizing wildlife. However, such restrictions might stimulate research and development into humane non-chemical devices, such as high-voltage, low-amperage restraint or very sophisticated (and expensive) traps.

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Table 17.1. Recommended doses of immobilizing drugs for selected species of furbearers (and potential non-target wildlife species that may be inadvertently captured during furbearer-related capture activities). The order of listing does not imply preference of drug regimens. Butorphanol-azaperone-medetomidine (BAM) doses are based on commercial kits consisting of premixed BAM (plus 25 mg/ml azaperone, 50 mg/ml naltrexone). Atipamezole is the preferred antagonist for all alpha-2 adrenoceptor agonists; it can be administered at a ratio of 1 mg atipamezole to 10 mg of xylazine administered.

Family or group Species	Dose and drug
Carnivora	
American badger (*Taxidea taxus*)	(a) 4.4 mg/kg tiletamine-zolazepam (b) 15 mg/kg ketamine plus 1 mg/kg xylazine (c) 0.4 ml BAM; antagonize with 0.8 ml atipamezole plus 0.5 ml naltrexone
American hog-nosed skunk (*Conepatus leuconotus*)	(a) 10 mg/kg tiletamine-zolazepam (b) 15 mg/kg ketamine plus 0.2 mg/kg acepromazine
American marten (*Martes americana*) Pacific marten (*Martes caurina*)	(a) 3 mg/kg tiletamine-zolazepam plus 2 mg/kg xylazine (b) 18 mg/kg ketamine plus 1.6 mg/kg xylazine (c) 10 mg/kg ketamine plus 0.2 mg/kg medetomidine; antagonize with 1 mg/kg atipamezole
American mink (*Neogale vison*)	(a) 15 mg/kg tiletamine-zolazepam (b) 40 mg/kg ketamine plus 1 mg/kg xylazine (c) 5 mg/kg ketamine plus 0.1 mg/kg medetomidine; antagonize with 0.5 mg/kg atipamezole
Arctic fox (*Vulpes lagopus*)	(a) 2.5 mg/kg ketamine plus 0.05 mg/kg medetomidine; antagonize with 0.25 mg/kg atipamezole (b) 0.1 ml BAM; antagonize with 0.2 ml atipamezole plus 0.5 ml naltrexone (c) 10 mg/kg tiletamine-zolazepam
Black-footed ferret (*Mustela nigripes*)	(a) 5 mg/kg ketamine plus 0.1 mg/kg medetomidine; antagonize with 0.5 mg/kg atipamezole (b) 25 mg/kg ketamine plus 2 mg/kg xylazine (c) 15 mg/kg tiletamine-zolazepam
Bobcat (*Lynx rufus*)	(a) 10 mg/kg tiletamine-zolazepam (b) 10 mg/kg ketamine plus 1.5 mg/kg xylazine (c) 0.3 ml BAM; antagonize with 0.5 ml atipamezole plus 0.5 ml naltrexone

Table 17.1. Continued.

Family or group Species	Dose and drug
Canada lynx (*Lynx canadensis*)	(a) 5 mg/kg tiletamine-zolazepam (b) 10 mg/kg ketamine plus 2 mg/kg xylazine (c) 5 mg/kg ketamine plus 0.2 mg/kg medetomidine; antagonize with 1 mg/kg atipamezole
Coyote (*Canis latrans*)	(a) 10 mg/kg tiletamine-zolazepam (b) 4 mg/kg ketamine plus 2 mg/kg xylazine; antagonize with 0.2 mg/kg atipamezole (c) 0.2 ml BAM; antagonize with 0.4 ml atipamezole plus 0.5 ml naltrexone
Eastern spotted skunk (*Spilogale putorius*)	(a) 10 mg/kg tiletamine-zolazepam (b) 16 mg/kg ketamine plus 8 mg/kg xylazine; antagonize with 0.8 mg/kg atipamezole (c) 15 mg/kg ketamine plus 0.2 mg/kg acepromazine
Gray fox (*Urocyon cinereoargenteus*)	(a) 9 mg/kg tiletamine-zolazepam (b) 15 mg/kg ketamine plus 3 mg/kg xylazine; antagonize with 0.3 mg/kg atipamezole
Gray wolf (*Canis lupus*)	(a) 4 mg/kg ketamine plus 0.08 mg/kg medetomidine; antagonize with 0.4 mg/kg atipamezole (b) 10 mg/kg ketamine plus 2 mg/kg xylazine; antagonize with 0.2 mg/kg atipamezole (c) 0.5 ml BAM; antagonize with 1 ml atipamezole plus 0.5 ml naltrexone (d) 10 mg/kg tiletamine-zolazepam
Hooded skunk (*Mephitis macroura*)	(a) 10 mg/kg tiletamine-zolazepam (b) 15 mg/kg ketamine plus 0.2 mg/kg acepromazine
Island fox (*Urocyon littoralis*)	(a) 9 mg/kg tiletamine-zolazepam (b) 15 mg/kg ketamine plus 3 mg/kg xylazine
Jaguarundi (*Herpailurus yagouaroundi*)	(a) 6.6 mg/kg tiletamine-zolazepam (b) 15 mg/kg ketamine plus 1 mg/kg xylazine
Kit fox (*Vulpes macrotis*)	(a) 10 mg/kg tiletamine-zolazepam (b) 20 mg/kg ketamine plus 0.2 mg/kg acepromazine
Least weasel (*Mustela nivalis*)	(a) 5 mg/kg ketamine plus 0.1 mg/kg medetomidine; antagonize with 0.5 mg/kg atipamezole (b) 25 mg/kg ketamine plus 2 mg/kg xylazine
Long-tailed weasel (*Neogale frenata*)	(a) 5 mg/kg ketamine plus 0.1 mg/kg medetomidine; antagonize with 0.5 mg/kg atipamezole (b) 25 mg/kg ketamine plus 2 mg/kg xylazine
Margay (*Leopardus wiedii*)	(a) 8.8 mg/kg tiletamine-zolazepam (b) 15 mg/kg ketamine plus 1 mg/kg xylazine
North American river otter (*Lontra canadensis*)	(a) 4 mg/kg ketamine plus 0.04 mg/kg medetomidine; antagonize with 0.2 mg/kg atipamezole (b) 4 mg/kg tiletamine-zolazepam (c) 7.5 mg/kg ketamine plus 1.5 mg/kg xylazine; antagonize with 0.15 mg/kg atipamezole
Northern raccoon (*Procyon lotor*)	(a) 5.5 mg/kg ketamine plus 0.055 mg/kg medetomidine; antagonize with 0.375 mg/kg atipamezole (b) 20 mg/kg ketamine plus 4 mg/kg xylazine; antagonize with 0.4 mg/kg atipamezole (c) 0.2 ml BAM; antagonize with 0.4 ml atipamezole plus 0.5 ml naltrexone (d) 3 mg/kg tiletamine-zolazepam
Ocelot (*Leopardus pardalis*)	(a) 5 mg/kg tiletamine-zolazepam (b) 15 mg/kg ketamine plus 1 mg/kg xylazine
Red fox (*Vulpes vulpes*)	(a) 2 mg/kg ketamine plus 0.08 mg/kg medetomidine; antagonize with 0.4 mg/kg atipamezole (b) 10 mg/kg tiletamine-zolazepam (c) 20 mg/kg ketamine plus 1 mg/kg xylazine; antagonize with 0.1 mg/kg atipamezole (d) 0.1 ml BAM; antagonize with 0.2 ml atipamezole plus 0.5 ml naltrexone
Ringtail (*Bassariscus astutus*)	(a) 10 mg/kg tiletamine-zolazepam (b) 15 mg/kg ketamine plus 0.2 mg/kg acepromazine

Table 17.1. Continued.

Family or group Species	Dose and drug
Sea otter (*Enhydra lutris*)	(a) 0.3 mg/kg fentanyl plus 0.1 mg/kg midazolam; antagonize with 0.6 mg/kg naltrexone
Short-tailed weasel (*Mustela erminea*)	(a) 5 mg/kg ketamine plus 0.1 mg/kg medetomidine; antagonize with 0.5 mg/kg atipamezole (b) 25 mg/kg ketamine plus 2 mg/kg xylazine
Striped skunk (*Mephitis mephitis*)	(a) 10 mg/kg tiletamine-zolazepam (b) 15 mg/kg ketamine plus 0.2 mg/kg acepromazine
Swift fox (*Vulpes velox*)	(a) 10 mg/kg tiletamine-zolazepam (b) 10 mg/kg ketamine plus 1 mg/kg xylazine; antagonize with 0.1 mg/kg atipamezole
Virginia opossum (*Didelphis virginiana*)	(a) 15 mg/kg tiletamine-zolazepam (b) 10 mg/kg ketamine plus 2 mg/kg xylazine (c) 10 mg/kg ketamine plus 0.1 mg/kg medetomidine; antagonize with 0.5 mg/kg atipamezole
Western spotted skunk (*Spilogale gracilis*)	(a) 10 mg/kg tiletamine-zolazepam (b) 16 mg/kg ketamine plus 8mg/kg xylazine; antagonize with 0.8 mg/kg atipamezole
White-nosed coati (*Nasua narica*)	(a) 8 mg/kg tiletamine-zolazepam (b) 20 mg/kg ketamine plus 1 mg/kg xylazine
Wolverine (*Gulo gulo*)	(a) 7 mg/kg ketamine plus 0.3 mg/kg medetomidine; antagonize with 1.5 mg/kg atipamezole (b) 20 mg/kg ketamine plus 0.2 mg/kg acepromazine
Rodentia	
American red squirrel (*Tamiasciurus hudsonicus*)	(a) 20 mg/kg ketamine plus 1mg/kg xylazine
Muskrat (*Ondatra zibethicus*)	(a) 50 mg/kg ketamine plus 5 mg/kg xylazine
North American beaver (*Castor canadensis*)	(a) 10 mg/kg ketamine plus 1 mg/kg xylazine (b) 5 mg/kg tiletamine-zolazepam (c) 0.4 ml BAM; antagonize with 0.8 ml atipamezole plus 0.5 ml naltrexone
North American porcupine (*Erethizon dorsatum*)	(a) 10 mg/kg tiletamine-zolazepam (b) 5 mg/kg ketamine plus 2 mg/kg xylazine; antagonize with 0.2 mg/kg atipamezole (c) 0.1 ml BAM; antagonize with 0.2 ml atipamezole plus 0.5 ml naltrexone
Nutria (*Myocastor coypus*)	(a) 5 mg/kg tiletamine-zolazepam (b) 4 mg/kg ketamine plus 0.5 mg/kg xylazine
Potential non-target species	
American black bear (*Ursus americanus*)	(a) 7 mg/kg tiletamine-zolazepam (b) 4.4 mg/kg ketamine plus 2 mg/kg xylzine; antagonize with 0.2 mg/kg atipamezole (c) 1.5 mg/kg ketamine plus 0.04 mg/kg medetomidine; antagonize with 0.2 mg/kg atipamezole (d) 1 ml/46 kg BAM; antagonize with 1 mg/kg atipamezole plus 0.25 mg/kg naltrexone
Mountain lion (*Puma concolor*)	(a) 0.1 ml/50 kg BAM; antagonize with 1 ml/50 kg atipamezole plus 0.5 ml naltrexone (b) 2.5 mg/kg ketamine plus 0.07 mg/kg medetomidine; antagonize with 0.35 mg/kg atipamezole (c) 5 mg/kg tiletamine-zolazepam plus 1 mg/kg xylazine; antagonize with 0.1 mg/kg atipamezole (d) 10 mg/kg ketamine plus 2 mg/kg xylazine; antagonize with 0.2 mg/kg atipamezole
White-tailed deer (*Odocoileus virginianus*)	(a) 2 ml BAM; antagonize with 4 ml atipamezole plus 0.5 ml naltrexone (b) 4.4 mg/kg tiletamine-zolazepam plus 2.2 mg/kg xylazine; antagonize with 0.22 mg/kg atipamezole (c) 7.5 mg/kg ketamine plus 1.5 mg/kg xylazine; antagonize with 0.15 mg/kg atipamezole (d) 2.5 mg/kg ketamine plus 0.1 mg/kg medetomidine; antagonize with 0.5 mg/kg atipamezole

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